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RESEARCH ARTICLE

Antibody testing for COVID-19: A report from the National COVID Scientific Advisory Panel [version 1; peer review: 2 approved]

Emily R. Adams¹, Mark Ainsworth², Rekha Anand³, Monique I. Andersson ², Kathryn Auckland⁴, J. Kenneth Baillie⁵, Eleanor Barnes^{2,4}, Sally Beer ², John I. Bell⁴, Tamsin Berry⁶, Sagida Bibi⁷, Miles Carroll^{4,8}, Senthil K. Chinnakannan ⁴, Elizabeth Clutterbuck⁷, Richard J. Cornall ^{2,4}, Derrick W. Crook ^{2,4}, Thushan de Silva⁹, Wanwisa Dejnirattisai⁴, Kate E. Dingle ⁴, Christina Dold⁷, Alexis Espinosa², David W. Eyre^{2,4}, Helen Farmer⁶, Maria Fernandez Mendoza², Dominique Georgiou², Sarah J. Hoosdally⁴, Alastair Hunter¹⁰, Katie Jefferey ², Dominic F. Kelly^{2,7}, Paul Klenerman^{2,4}, Julian Knight^{2,4}, Clarice Knowles⁶, Andrew J. Kwok⁴, Ullrich Leuschner¹¹, Robert Levin¹², Chang Liu⁴, César López-Camacho ⁴, Jose Martinez², Philippa C. Matthews ^{2,4}, Hannah McGivern ¹³, Alexander J. Mentzer ^{2,4}, Jonathan Milton¹³, Juthathip Mongkolsapaya⁴, Shona C. Moore¹⁴, Marta S. Oliveira¹³, Fiona Pereira¹⁵, Elena Perez², Timothy Peto^{2,4}, Rutger J. Ploeg ^{2,13}, Andrew Pollard ^{2,7}, Tessa Prince ¹⁴, David J. Roberts¹¹, Justine K. Rudkin ⁴, Veronica Sanchez², Gavin R. Screaton⁴, Malcolm G. Semple ^{14,16}, Jose Slon-Campos⁴, Donal T. Skelly ^{2,17}, Elliot Nathan Smith⁶, Alberto Sobrinodiaz², Julie Staves², David I. Stuart ^{4,18}, Piyada Supasa⁴, Tomas Surik¹³, Hannah Thraves², Pat Tsang¹¹, Lance Turtle ^{14,19}, A. Sarah Walker⁴, Beibei Wang⁴, Charlotte Washington³, Nicholas Watkins²⁰, James Whitehouse⁶, National COVID Testing Scientific Advisory Panel

¹Liverpool School of Tropical Medicine, Liverpool, L3 5QA, UK

²Oxford University Hospitals NHS Foundation Trust, Oxford, OX3 9DU, UK

³NHS Blood and Transplant Birmingham, Vincent Drive, Birmingham, B15 2SG, UK

⁴Nuffield Department of Medicine and NIHR Oxford Biomedical Research Centre, University of Oxford, Oxford, OX3 9DU, UK

⁵Roslin Institute, University of Edinburgh, Edinburgh, EH25 9RJ, UK

⁶Department of Health and Social Care, UK Government, London, UK

⁷Department of Paediatrics, Oxford Vaccine Group, University of Oxford, Oxford, OX3 7LE, UK

⁸Public Health England, Porton Down, Salisbury, SP4 0JG, UK

⁹Department of Infection, Immunity and Cardiovascular Disease, University of Sheffield, Sheffield, S10 2RX, UK

¹⁰NHS Blood and Transplant Basildon, Burnt Mills Industrial Estate, Basildon, SS13 1FH, UK

¹¹NHS Blood and Transplant Oxford, John Radcliffe Hospital, Oxford, UK

¹²Worthing Hospital, Worthing, BN11 2DH, UK

¹³Nuffield Department of Surgical Sciences, University of Oxford, Oxford, OX3 9DU, UK

¹⁴NIHR Health Protection Research Unit in Emerging and Zoonotic Infections, Faculty of Health and Life Sciences, University of Liverpool, Liverpool, UK

¹⁵Imperial College London, London, SW7 2AZ, UK

¹⁶Alder Hey Children's Hospital, Liverpool, UK

¹⁷Nuffield Department of Clinical Neurosciences, University of Oxford, Oxford, OX3 9DU, UK

¹⁸Diamond Light Source, Harwell Science and Innovation Campus, Didcot, OX11 0DE, UK

¹⁹Tropical & Infectious Disease Unit, Royal Liverpool University Hospital (member of Liverpool Health Partners), Liverpool, L7 8XP, UK

²⁰NHS Blood and Transplant Cambridge, Long Road, Cambridge, CB2 0PT, UK

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Abstract

Background: The COVID-19 pandemic caused >1 million infections during January-March 2020. There is an urgent need for reliable antibody detection approaches to support diagnosis, vaccine development, safe release of individuals from quarantine, and population lock-down exit strategies. We set out to evaluate the performance of ELISA and lateral flow immunoassay (LFIA) devices.

Methods: We tested plasma for COVID (severe acute respiratory syndrome coronavirus 2; SARS-CoV-2) IgM and IgG antibodies by ELISA and using nine different LFIA devices. We used a panel of plasma samples from individuals who have had confirmed COVID infection based on a PCR result (n=40), and pre-pandemic negative control samples banked in the UK prior to December-2019 (n=142).

Results: ELISA detected IgM or IgG in 34/40 individuals with a confirmed history of COVID infection (sensitivity 85%, 95%CI 70-94%), vs. 0/50 pre-pandemic controls (specificity 100% [95%CI 93-100%]). IgG levels were detected in 31/31 COVID-positive individuals tested ≥ 10 days after symptom onset (sensitivity 100%, 95%CI 89-100%). IgG titres rose during the 3 weeks post symptom onset and began to fall by 8 weeks, but remained above the detection threshold. Point estimates for the sensitivity of LFIA devices ranged from 55-70% versus RT-PCR and 65-85% versus ELISA, with specificity 95-100% and 93-100% respectively. Within the limits of the study size, the performance of most LFIA devices was similar.

Conclusions: Currently available commercial LFIA devices do not perform sufficiently well for individual patient applications. However, ELISA can be calibrated to be specific for detecting and quantifying SARS-CoV-2 IgM and IgG and is highly sensitive for IgG from 10 days following first symptoms.

Keywords

COVID-19, SARS-CoV-2, serology, IgG, IgM, antibodies, immunoassay, ELISA, lateral flow, exposure, epidemiology

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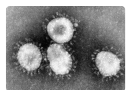
11 Jun 2020


report


report

1. **Kim Tien Ng** , National University Health System (NUHS), National University of Singapore, Singapore, Singapore
- Yee Joo Tan** , National University Health System (NUHS), National University of Singapore & Institute of Molecular and Cell Biology (IMCB), A*STAR (Agency for Science, Technology and Research), Singapore, Singapore
2. **Muki Shey** , University of Cape Town, Cape Town, South Africa

Any reports and responses or comments on the article can be found at the end of the article.



This article is included in the [Coronavirus \(COVID-19\)](#) collection.

Corresponding authors: Derrick W. Crook (derrick.crook@ndcls.ox.ac.uk), Gavin R. Screaton (gavin.screaton@medsci.ox.ac.uk)

Author roles: **Adams ER:** Data Curation, Resources; **Ainsworth M:** Data Curation, Resources; **Anand R:** Data Curation, Resources; **Andersson MI:** Data Curation, Resources; **Auckland K:** Data Curation; **Baillie JK:** Conceptualization, Data Curation, Methodology, Resources; **Barnes E:** Supervision; **Beer S:** Data Curation, Resources; **Bell JI:** Conceptualization, Methodology, Supervision; **Berry T:** Conceptualization, Supervision; **Bibi S:** Conceptualization; **Carroll M:** Supervision; **Chinnakannan SK:** Data Curation; **Clutterbuck E:** Data Curation, Resources; **Cornall RJ:** Supervision; **Crook DW:** Conceptualization, Methodology, Supervision, Writing – Original Draft Preparation, Writing – Review & Editing; **de Silva T:** Data Curation, Resources; **Dejnirattisai W:** Data Curation; **Dingle KE:** Data Curation; **Dold C:** Data Curation; **Espinosa A:** Data Curation, Resources; **Eyre DW:** Conceptualization, Formal Analysis, Methodology, Supervision, Writing – Original Draft Preparation, Writing – Review & Editing; **Farmer H:** Supervision; **Fernandez Mendoza M:** Data Curation, Resources; **Georgiou D:** Data Curation, Resources; **Hoosdally SJ:** Supervision; **Hunter A:** Data Curation, Resources; **Jefferey K:** Data Curation, Resources; **Kelly DF:** Resources, Supervision; **Klenerman P:** Supervision; **Knight J:** Supervision; **Knowles C:** Supervision; **Kwok AJ:** Data Curation; **Leuschner U:** Data Curation, Resources; **Levin R:** Data Curation, Resources; **Liu C:** Data Curation; **López-Camacho C:** Data Curation; **Martinez J:** Data Curation, Resources; **Matthews PC:** Conceptualization, Formal Analysis, Methodology, Supervision, Writing – Original Draft Preparation, Writing – Review & Editing; **McGivern H:** Data Curation, Resources; **Mentzer AJ:** Conceptualization, Data Curation, Methodology, Resources, Supervision; **Milton J:** Data Curation, Resources; **Mongkolsapaya J:** Data Curation; **Moore SC:** Data Curation, Resources; **Oliveira MS:** Data Curation, Resources; **Pereira F:** Supervision; **Perez E:** Data Curation, Resources; **Peto T:** Conceptualization, Formal Analysis, Methodology, Writing – Original Draft Preparation, Writing – Review & Editing; **Ploeg RJ:** Conceptualization, Supervision; **Pollard A:** Data Curation, Resources; **Prince T:** Data Curation, Resources; **Roberts DJ:** Data Curation, Resources, Supervision; **Rudkin JK:** Data Curation; **Sanchez V:** Data Curation, Resources; **Screaton GR:** Conceptualization, Data Curation, Methodology, Supervision; **Semple MG:** Conceptualization, Data Curation, Methodology, Resources; **Slon-Campos J:** Data Curation, Resources; **Skelly DT:** Data Curation, Resources; **Smith EN:** Supervision; **Sobrinodiaz A:** Data Curation, Resources; **Staves J:** Conceptualization; **Stuart DI:** Data Curation, Supervision; **Supasa P:** Data Curation; **Surik T:** Data Curation, Resources; **Thraves H:** Data Curation, Resources; **Tsang P:** Data Curation, Resources; **Turtle L:** Data Curation, Resources; **Walker AS:** Conceptualization, Formal Analysis, Methodology, Writing – Original Draft Preparation, Writing – Review & Editing; **Wang B:** Data Curation; **Washington C:** Data Curation, Resources; **Watkins N:** Data Curation, Resources; **Whitehouse J:** Supervision;

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Introduction

The first cases of infection with a novel coronavirus (severe acute respiratory syndrome coronavirus 2; SARS-CoV-2) causing coronavirus infectious disease (COVID) emerged in Wuhan, China on December 31st, 2019¹. Despite intensive containment efforts, there was rapid international spread and three months later, there had been over 1 million confirmed infections and 60,000 reported deaths². Containment efforts have relied heavily on population quarantine ('lock-down') measures to restrict movement and reduce individual contacts^{3,4}. To develop public health strategies for exit from lock-down, diagnostic testing urgently needs to be scaled-up, including both mass screening and screening of specific high-risk groups (contacts of confirmed cases, and healthcare workers and their families), in parallel with collecting data on recent and past infection at individual and population levels².

Laboratory diagnosis of infection has mostly been based on real-time RT-PCR, typically targeting the viral RNA-dependent RNA polymerase (RdRp) or nucleocapsid (N) genes using swabs collected from the upper respiratory tract^{5,6}. This requires specialist equipment, skilled laboratory staff and PCR reagents, creating diagnostic delays. RT-PCR from upper respiratory tract swabs may also be falsely negative due to quality or timing; viral loads in upper respiratory tract secretions peak in the first week of symptoms⁷, but may have declined below the limit of detection in those presenting later⁸. In individuals who have recovered, RT-PCR provides no information about prior exposure or immunity.

In contrast, assays that reliably detect antibody responses specific to SARS-CoV-2 could contribute to diagnosis of acute infection (via rises in IgM and IgG levels) and to identifying those infected with or without symptoms and recovered (via persisting IgG)⁹. Receptor-mediated viral entry to host cells occurs through interactions between the unique and highly-conserved viral spike (S) glycoprotein and the ACE2 cell receptor¹⁰. This S protein is the primary target of specific neutralising antibodies, and current SARS-CoV-2 serology assays therefore typically seek to identify these antibodies (Figure 1A–C). Rapid lateral flow immunoassay (LFIA) devices provide a quick, point-of-care approach to antibody testing. A sensitive and specific antibody assay could directly contribute to early identification and isolation of cases, address unknowns regarding the extent of infection to inform mathematical models and support individual or population-level release from lock-down. Laboratory-based ELISA platforms have also been evaluated as an approach to detection and quantification of SARS-CoV-2 antibodies¹¹.

However, before either laboratory assays or LFIA devices can be widely deployed, their performance needs to be carefully evaluated (Figure 1D, E)¹². We therefore compared a novel laboratory-based ELISA assay with nine commercially-available LFIA devices using samples from patients with RT-PCR-confirmed infection, and negative pre-pandemic samples.

Methods

Research reporting

Samples. A total of 142 plasma samples designated seronegative for SARS-CoV-2 were collected from adults (≥ 18 years)

in the UK before December 2019 (*Underlying data*, Table S1, including demographic details¹³) from three ethically approved sources: healthy blood donors, organ donors on ICU following cerebral injury and healthy volunteers from a vaccine study.

In total, 40 plasma samples were collected from adults positive for SARS-CoV-2 by RT-PCR from an upper respiratory tract (nose/throat) swab tested in accredited laboratories (*Underlying data*, Table S1¹³). Acute (≤ 28 days from symptom onset) and convalescent samples (>28 days) were included to optimise detection of SARS-CoV-2 specific IgM and IgG respectively (Figure 1B). Acute samples were collected from patients a median 10 (range 4–27) days from symptom onset ($n=16$), and from recovering healthcare workers median 13 [range 8–19] days after first symptoms; ($n=6$). Convalescent samples were collected from adults a median 48 [range 31–62] days after symptom onset and/or date of positive throat swab ($n=18$). Further sample details are provided in *Extended data*, Supplementary Material¹³.

Cases were classified following WHO criteria as critical (respiratory failure, septic shock, and/or multiple organ dysfunction/failure); severe (dyspnoea, respiratory frequency ≥ 30 /minute, blood oxygen saturation $\leq 93\%$, $\text{PaO}_2/\text{FiO}_2$ ratio <300 , and/or lung infiltrates $>50\%$ of the lung fields within 24–48 hours); or otherwise mild¹⁴. Among 22 acute cases, 9 were critical, 4 severe and 9 mild. All but one convalescent individual had mild disease; the other was asymptomatic and screened during enhanced contact tracing.

ELISA

We developed a novel ELISA targeting the SARS-CoV-2 spike protein. Recombinant SARS-CoV-2 trimeric spike protein was constructed as described¹⁵, using mammalian codon optimized SARS2 Spike (1–1208, Genbank accession [MN908947](#)) with a GSAS substitution at the furin cleavage site (aa 682–685) and double proline substitution at aa 986–987. The C-terminal was followed by T4 fibrin motif, an HRV3C protease cleavage site, a TwinStrep Tag and an 8-HisTag. The gene was cloned into a pHLsec and expressed in 293T cells. The HIS trap HP column (cat no 17524701; Cytiva) was used to purify the recombinant S protein.

We used ELISA to detect antibodies to the S protein. MAXISORP immunoplates (442404; NUNC) were coated with StrepMAB-Classic (2-1507-001; ibi). Plates were blocked with 2% skimmed milk in PBS for one hour and then incubated with 0.125 μg of soluble trimeric SARS-CoV-2 trimeric S protein or 2% skimmed milk in phosphate buffered saline. After one hour, plasma was added at 1:50 dilution, followed by ALP-conjugated anti-human IgG (A9544, RRID:AB_258459; Sigma) at 1:10,000 dilution or ALP-conjugated anti-human IgM (A9794, RRID:AB_258474; Sigma) at 1:5,000 dilution. The reaction was developed by the addition of PNPP substrate and stopped with 1.0 M NaOH. The absorbance was measured at 405nm after 90 minutes, and a final optical density (OD) value was calculated by subtracting the background (skimmed milk) from the test value. The ELISA assay takes 5–6 hours to perform with an experienced operator being able to process up to five 96-well plates (480 samples including relevant controls).

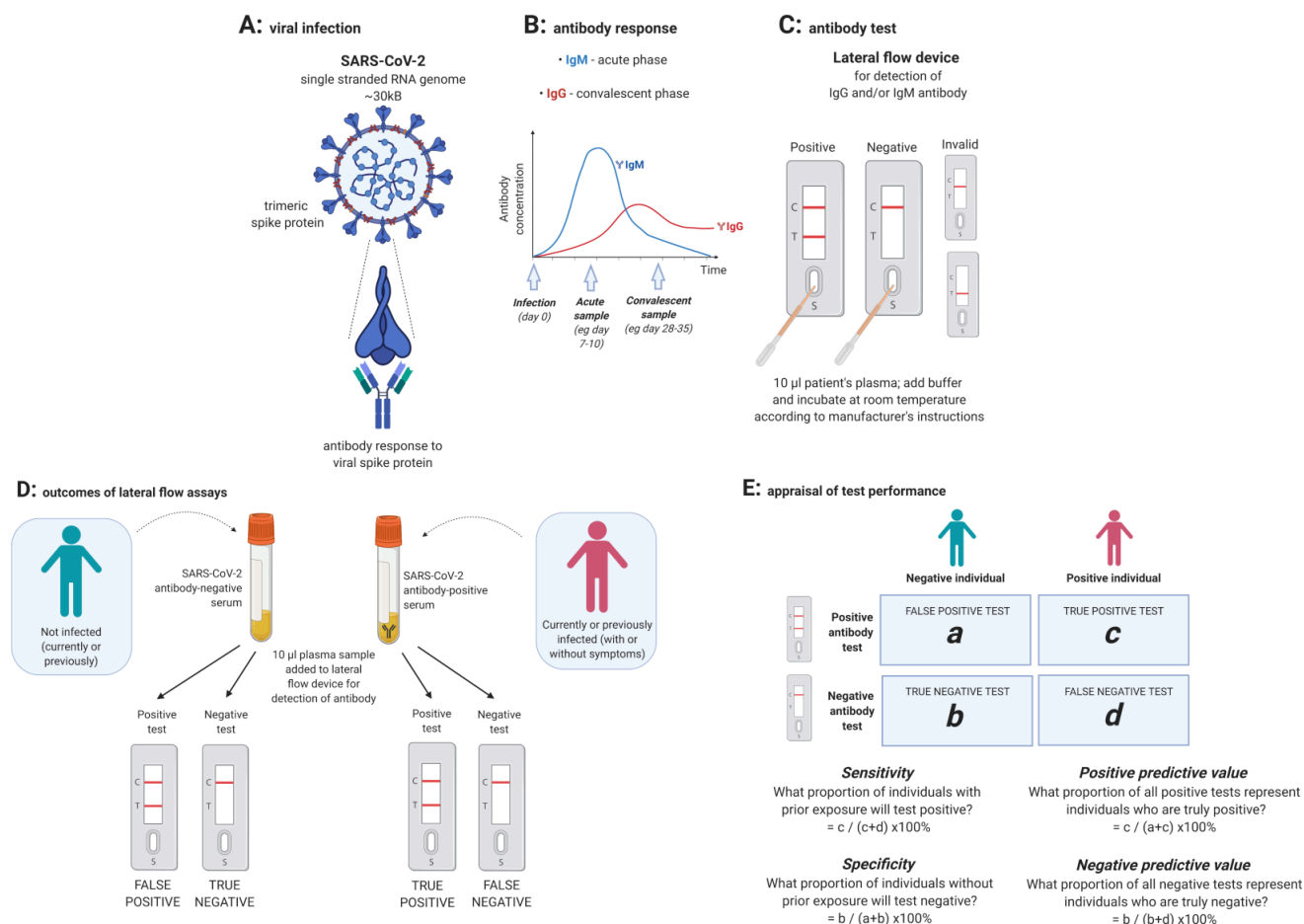


Figure 1. Cartoon to illustrate the generation of IgM and IgG antibodies to SARS nCoV-2 and detection of antibodies by a lateral flow device. (A) *In vivo* generation of antibodies to the trimeric SARS-CoV-2 spike protein. **(B)** Projected change in titres of specific IgM and IgG over time following infection, with arrows indicating typical time frames for collection of acute and convalescent samples. **(C)** *Ex vivo* detection of IgG and/or IgM using a lateral flow immunoassay (LFIA): S= sample well, T=test antibody; C=control. Diagram shows a positive sample on the left, with positive lines at both C and T, and a negative test on the right with a line present only at C. Any other combination of lines renders the test invalid. Some devices have two test lines, for separate detection of anti-SARS-CoV-2-IgG and -IgM. Assays variably suggest use of plasma, serum and/or whole blood. **(D)** Outcomes of testing negative and positive samples using LFIA. **(E)** Calculation of sensitivity, specificity, positive and negative predictive value of a test. Image created with BioRender.com; exported under a paid subscription.

LFIA

We tested LFIA devices designed to detect IgM, IgG or total antibodies to SARS-CoV-2 produced by nine manufacturers short-listed as a testing priority by the UK Government Department of Health and Social Care (DHSC), based on appraisals of device provenance and available performance data. Individual manufacturers did not approve release of device-level data, so device names are anonymised.

Testing was performed in strict accordance with the manufacturer's instructions for each device. Typically, this involved adding 5–20 μ l of plasma to the sample well, and 80–100 μ l of manufacturer's buffer to an adjacent well, followed by incubation at room temperature for 10–15 minutes. The result was based on the appearance of coloured bands, designated as positive

(control and test bands present), negative (control band only), or invalid (no band, absent control band, or band in the wrong place) (Figure 1C).

We recorded results in real-time on a password-protected electronic database, using pseudonymised sample identifiers, capturing the read-out from the device (positive/negative/invalid), operator, device, device batch number, and a timestamped photograph of the device.

Testing protocol

We tested 90 samples using ELISA to quantify IgM and IgG antibody in plasma designated SARS-CoV-2 negative (n=50) and positive (n=40). All positive samples were included and an unstratified random sample of negative plasma from healthy blood

donors (n=23) and organ donors (n=27). We tested the nine different LFIA devices using between 39–165 individual plasma samples (8–23 and 31–142 samples designated SARS-CoV-2 positive and negative, respectively, Table S2¹³). Total numbers varied according to the number of devices supplied to the DHSC; samples were otherwise selected at random.

Statistical analysis

Analyses were conducted using R (version 3.6.3) and Stata (version 15.1), with additional plots generated using GraphPad Prism (version 8.3.1). Binomial 95% confidence intervals (CI) were calculated for all proportions. The association between ELISA results and time since symptom onset, severity, need for hospital admission and age was estimated using multivariable linear regression, without variable selection. Non-linearity in relationships with continuous factors was included via natural cubic splines. Differences between LFIA devices were estimated using mixed effects logistic regression models, allowing for each device being tested on overlapping sample sets. Differences between devices were compared with Benjamini-Hochberg corrected p-value thresholds. (Further details in *Extended data*, Supplementary Material¹³.)

Ethical approval and role of the funding source

Our work was undertaken with ethical approval from the National Health Service Blood and Transplant (NHSBT) ethics, providing donor consent for plasma use; NIHR Biobank REC agreement (REC 13/NW/0017; IRAS 87824); International Severe Acute Respiratory and Emerging Infection Consortium ('ISARIC') approval by the South Central (Oxford C) Research Ethics Committee in England (Ref: 13/SC/0149), and Scotland A Research Ethics Committee in Scotland (Ref: 20/SS/0028). The UK Government DHSC selected the lateral flow devices for testing as described above. Otherwise, the funders had no role in study design or in the collection, analysis, and interpretation of data. Authors from DHSC contributed to writing of the report and in the decision to submit the paper for publication.

An earlier version of this article can be found on medRxiv (DOI: <https://doi.org/10.1101/2020.04.15.20066407>).

Results

Detection of SARS-CoV-2 IgM and IgG antibody by ELISA

The 40 positive (RT-PCR-confirmed SARS-CoV-2 infection) and 50 designated negative (pre-pandemic) plasma samples were tested by ELISA to characterise antibody profiles. Negative samples had median optical density (OD) for IgM of -0.0001 (arbitrary units) (range -0.14 to 0.06) and for IgG -0.01 (range -0.38 to 0.26). The median IgM reading in 40 positive samples was 0.18 (range -0.008 to 1.13; Kruskal-Wallis $p < 0.001$ vs. negative) and IgG median 3.0 (range -0.2 to 3.5; $p < 0.001$).

As safe individual release from lock-down is a major application for serological testing, we chose OD thresholds that maintained 100% specificity (95%CI 93–100%), while maximising sensitivity. Using thresholds of 0.07 for IgM and 0.4 for IgG (3 and 5 standard deviations above the negative mean, respectively;

Figure 2A, B), the IgG assay had 85% sensitivity (95%CI 70–94%; 34/40) vs. RT-PCR diagnosis. All six false negatives were from samples taken within 9 days of symptom onset (Figure 2D). IgG levels were detected in 31/31 RT-PCR-positive individuals tested ≥ 10 days after symptom onset (sensitivity 100%, 95%CI 89–100%). The IgM assay sensitivity was lower at 70% (95%CI 53–83%; 28/40). All IgG false-negatives were IgM-negative. No sample was IgM-positive and IgG-negative.

Considering the relationship between IgM and IgG titres and time since symptom onset (Figures 2C, D), univariable regression models showed IgG antibody titres rising over the first 3 weeks from symptom onset. The lower bound of the pointwise 95%CI for the mean expected titre crosses our OD threshold between days 6–7 (Figure 2D). However, given sampling variation, test performance is likely to be optimal from several days later. IgG titres fell during the second month after symptom onset but remained above the OD threshold. No temporal association was observed between IgM titres and time since symptom onset (Figure 2C). There was no evidence that SARS-CoV-2 severity, need for hospital admission or patient age were associated with IgG or IgM titres in multivariable models ($p > 0.1$, Table S3¹³).

Detection of SARS-CoV-2 antibodies by LFIA vs. RT-PCR

We first considered performance of the nine different LFIA devices using RT-PCR-confirmed cases as the reference standard (Table 1A and *Extended data*, Figure S1¹³) and considering any LFIA positive result (IgM, IgG or both) as positive. The LFIA devices achieved sensitivity ranging from 55% (95%CI 36–72%) to 70% (51–84%) and specificity from 95% (95%CI 86–99%) to 100% (94–100%). There was no evidence of differences between the devices in sensitivity ($p \geq 0.015$, cf. Benjamini-Hochberg $p = 0.0014$ threshold) or specificity ($p \geq 0.19$ for all devices with at least one false-positive test). Restricting to 31 samples collected ≥ 10 days post symptom-onset (all ELISA IgG-positive), LFIA sensitivity ranged from 61% (95%CI 39–80%) to 88% (68–97%) (*Extended data*, Table S4¹³).

Detection of SARS-CoV-2 antibodies by LFIA vs. ELISA

We also considered performance relative to ELISA (*Extended data*, Table S5, Figure S1¹³), because the LFIA devices target the same antibodies. We considered patients positive by this alternative standard if their IgG OD reading exceeded the threshold described above, since no samples were IgM-positive, IgG-negative). Sensitivity of antibody detection by LFIA ranged from 65% (95%CI 46–80%) to 85% (66–96%) and specificity from 93% (95%CI 83–98%) to 100% (94–100%); however, the device with the highest sensitivity had one of the lowest specificities (*Extended data*, Figure S1¹³). There was no evidence of differences in sensitivity ($p \geq 0.010$, cf. $p = 0.0014$ threshold) or specificity between devices ($p \geq 0.19$).

Of 50 designated negative samples tested by both ELISA and the nine different LFIA devices, nine separate samples generated at least one false-positive, on seven different LFIA devices (Figure 3). Four samples generating false-positive results did so on more than one LFIA device, despite the absence of

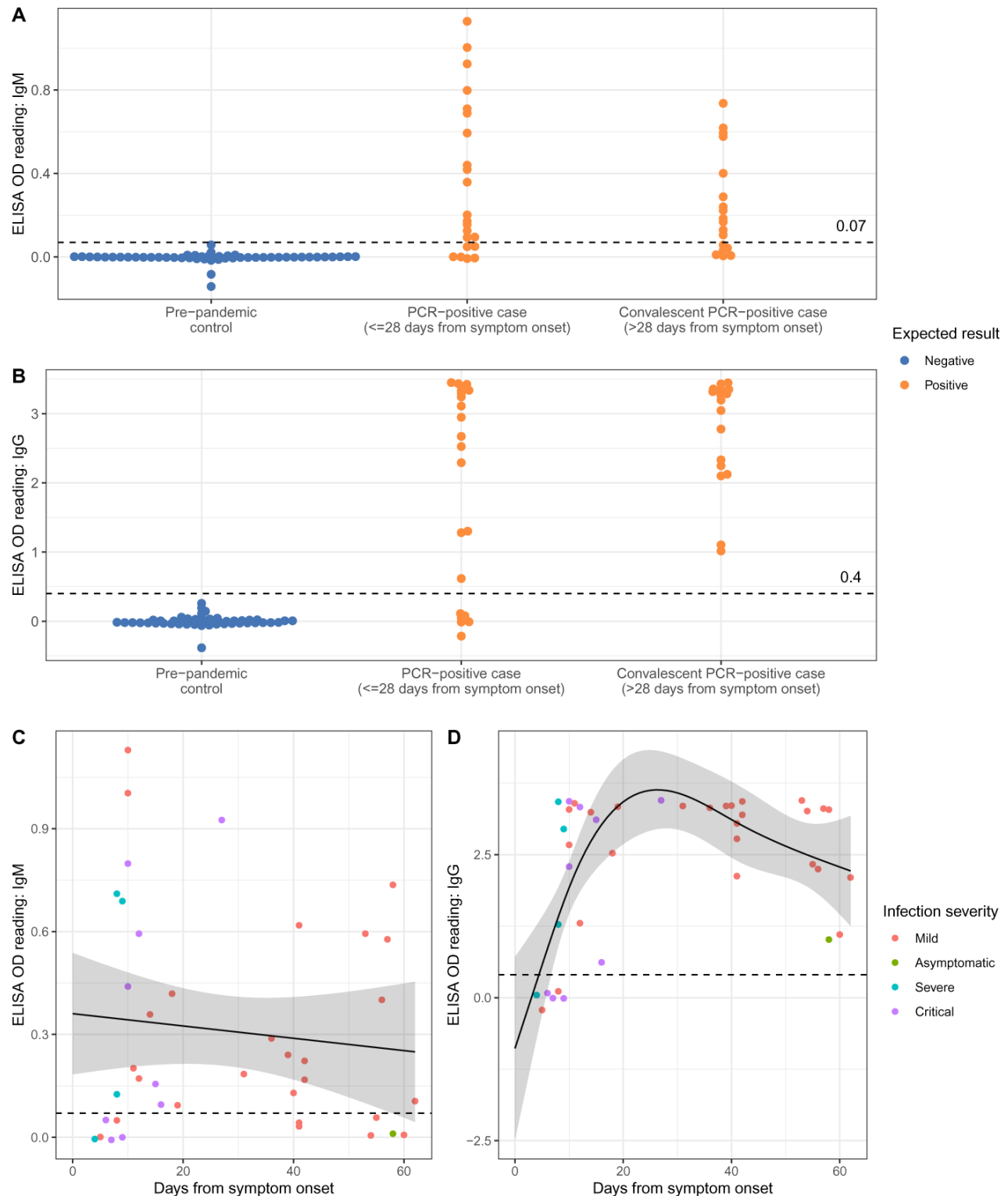


Figure 2. Results of testing 90 plasma samples for SARS-CoV-2 IgM and IgG by Enzyme linked Immunosorbent Assay (ELISA). (A) IgM readings for SARS-CoV-2 pre-pandemic plasma (designated negatives, shown in blue, $n=50$), and RT-PCR confirmed cases of SARS-CoV-2 infection (designated positives, shown in orange, $n=40$; divided into acute cases, $n=22$, and convalescent cases, $n=18$). Threshold of OD = 0.07 discriminates accurately between negative controls and convalescent sera. (B) IgG data shown for the same subgroups described for panel (A). A threshold of OD = 0.4 discriminates between designated negatives and positives. (C) IgM OD values plotted against the time post symptoms at which plasma was obtained. The line shows the mean OD value expected from a spline-based linear regression model, the ribbon indicates the pointwise 95% confidence interval. (D) IgG OD values plotted against the time post symptoms at which plasma was obtained. Coloured dots in panels C and D indicate disease severity. OD = optical density.

Table 1. Results of nine lateral flow immunoassays (LFIA) devices and an ELISA assay, tested with plasma classified as positive (RT-PCR positive) and negative (pre-pandemic). n=91–182 per LFIA device. Different manufacturers are designated 1–9. 95% confidence intervals (CI) are presented for each point estimate. Any LFIA positive result (IgM, IgG or both) was considered positive. ELISA positive samples were all positive for IgG, no sample was IgM-positive and IgG-negative.

Assay	RT-PCR positive		Pre-pandemic control		Sensitivity (95% CI)	Specificity (95% CI)
	True positive	False negative	True negative	False positive		
ELISA	34	6	50	0	85 (70,94)	100 (93,100)
1	18	15	60	0	55 (36,72)	100 (94,100)
2	23	15	90	1	61 (43,76)	99 (94,>99)
3	21	12	58	2	64 (45,80)	97 (88,>99)
4	25	13	59	1	66 (49,80)	98 (91,>99)
5	19	12	58	2	61 (42,78)	97 (91,>99)
6	20	11	59	1	65 (45,81)	98 (91,>99)
7	23	10	57	3	70 (51,84)	95 (86,>99)
8	18	14	60	0	56 (38,74)	100 (94,100)
9	22	18	138	4	55 (38,74)	97 (93,>99)

quantifiable IgM or IgG on ELISA, potentially suggesting a specific attribute of the sample causing a cross-reaction on certain LFIA platforms.

Of the 22 samples collected from RT-PCR positive patients in the acute setting, six fell below the ELISA detection threshold for IgM or IgG; two of these six were positive on LFIA testing, each on one (different) device. Of the remaining 16 acute samples (all ELISA IgG-positive), only nine were consistently positive across all nine LFIA devices. Due to limited availability of LFIA devices, fewer tests were performed on the 18 convalescent samples with available ELISA data, all with quantifiable IgG (Figure 2B, Figure 3A). Two had no antibody detected on any LFIA device, and only eight were consistently positive across all LFIA devices tested (between 1 and 9 devices tested per sample). Full metadata for results of ELISA and LFIA devices are available in *Underlying data*, Supplementary Table S6¹³.

Discussion

We here present the performance characteristics of a novel ELISA and nine selected LFIA devices for detecting SARS-CoV-2 IgM and IgG. Among 40 RT-PCR-confirmed positive patients, 85% had IgG detected by ELISA, including 100% patients tested ≥ 10 days after symptom onset. A panel of LFIA devices had sensitivity between 55 and 70% against the reference-standard RT-PCR, or 65–85% against ELISA, with specificity of 95–100% and 93–100%, respectively. These estimates come with wide confidence intervals due to constraints on the number of devices made available. Comparable results have been

obtained through a similar appraisal undertaken independently, in which specificity ranged from 84–100.0%, and the proportion of specimens testing positive increased over time from symptom onset, with >80% sensitivity achieved by some LFIA devices at later time points¹⁶. Our study, and these parallel data from another centre¹⁶, provide a benchmark against which to assess the performance of future antibody testing platforms, with the aim of guiding decisions about deploying antibody testing and informing the design of second-generation assays.

LFIA devices are cheap to manufacture, store and distribute, and could be used as a point-of-care test, offering an appealing approach to diagnostics and evaluating exposure, were adequate performance to be confirmed. A positive antibody test is currently regarded as a probable surrogate for immunity to reinfection. Secure confirmation of antibody status would therefore reduce anxiety, provide confidence to allow individuals to relax social distancing measures, and guide policy-makers in the staged release of population lock-down, potentially in tandem with digital approaches to contact tracing¹⁷. As a diagnostic tool, serology may have a role in combination with RT-PCR testing to improve sensitivity, particularly of cases presenting some time after symptom onset^{18,19}. Reproducible methods to detect and quantify vaccine-mediated antibodies are also crucial as COVID vaccines enter clinical trials.

Appropriate thresholds for sensitivity and specificity depend on the primary purpose of the test. For diagnosis in symptomatic patients, high sensitivity is required (generally $\geq 90\%$). Specificity

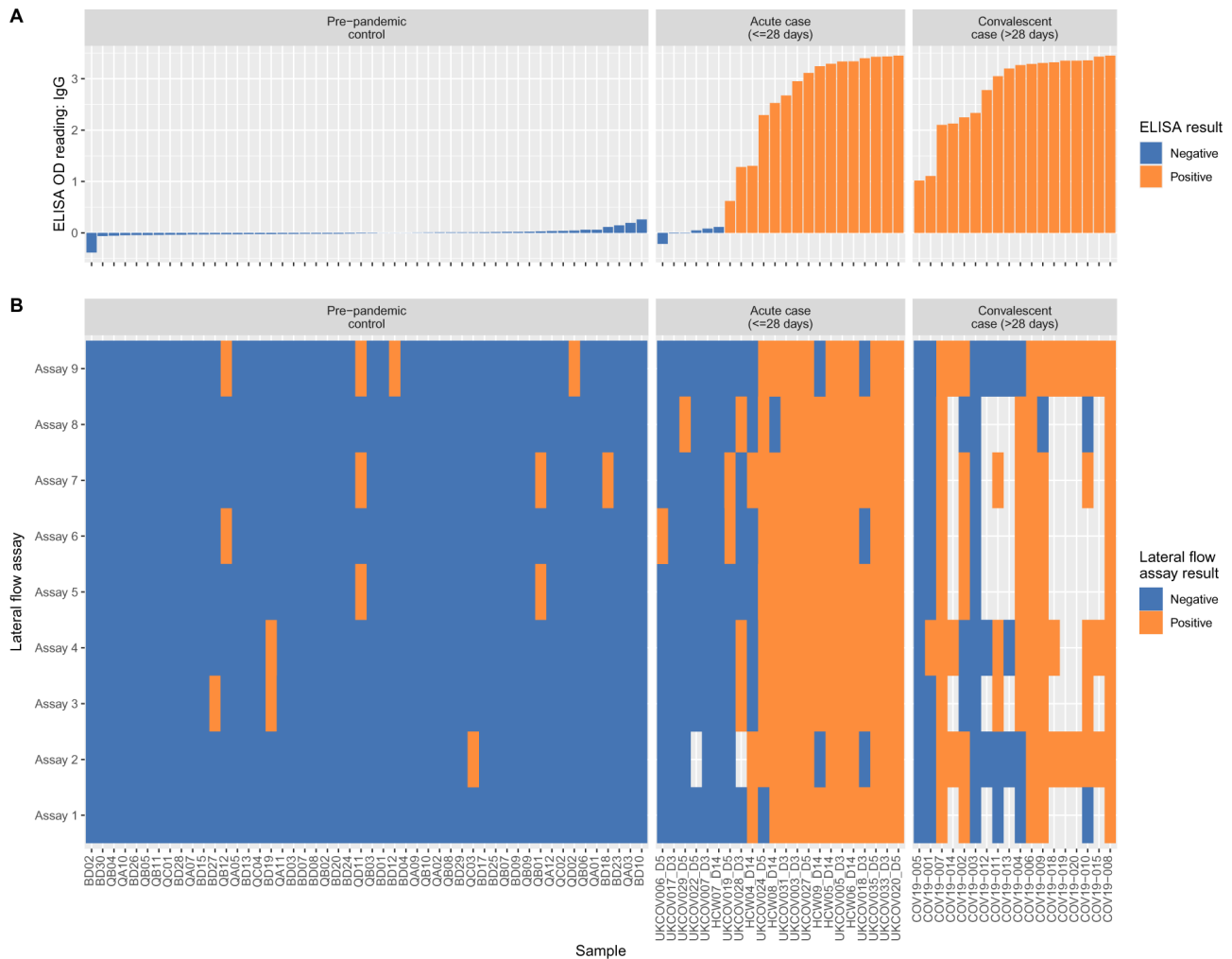


Figure 3. Comparison between ELISA and LFIA for SARS-CoV-2 designated negative and positive plasma. (A) Quantitative optical density (OD) readout from ELISA for IgG for designated negative plasma (n=50) and from individuals with RT-PCR confirmed infection (n=40, divided into acute and convalescent plasma). IgM results are shown in *Extended data*, Figure S2¹³. **(B)** Results from LFIA produced by nine manufacturers. Any positive test for IgG, IgM, both or total antibody is shown as positive; see *Extended data*, Figure S2 for more detailed breakdown¹³. Grey blocks indicate missing data as a result of insufficient devices to test all samples and one assay on one device with an invalid result. Samples in both panels are ranked from left to right by quantitation of IgG, as indicated in panel (A).

is less critical as some false-positives could be tolerated (provided other potential diagnoses are considered, and accepting that over-diagnosis causes unnecessary quarantine or hospital admission). However, if antibody tests were deployed as an individual-level approach to inform release from quarantine, then high specificity is essential, as false-positive results return non-immune individuals to risk of exposure. For this reason, the UK Medicines and Healthcare products Regulatory Agency has currently set a minimum 98% specificity threshold for LFIAs²⁰.

Appraisal of test performance should also consider the influence of population prevalence, acknowledging that this changes over time, geography and within different population groups. The potential risk of a test providing false reassurance and

release from lock-down of non-immune individuals can be considered as the proportion of all positive tests that are wrong. Based on the working 'best case' scenario of a LFIA test with 70% sensitivity and 98% specificity, the proportion of positive tests that are wrong is 35% at 5% population seroprevalence (19 false-positives/1000 tested), 10% at 20% seroprevalence (16 false-positives/1000) and 3% at 50% seroprevalence (10 false-positives/1000) (Figure 4).

More data are needed to investigate antibody-positivity as a correlate of protective immunity. Indeed, pre-existing IgG could enhance disease in some situations²¹, with animal data demonstrating that SARS-CoV-2 anti-spike IgG contributes to a proinflammatory response associated with lung injury in macaques²².

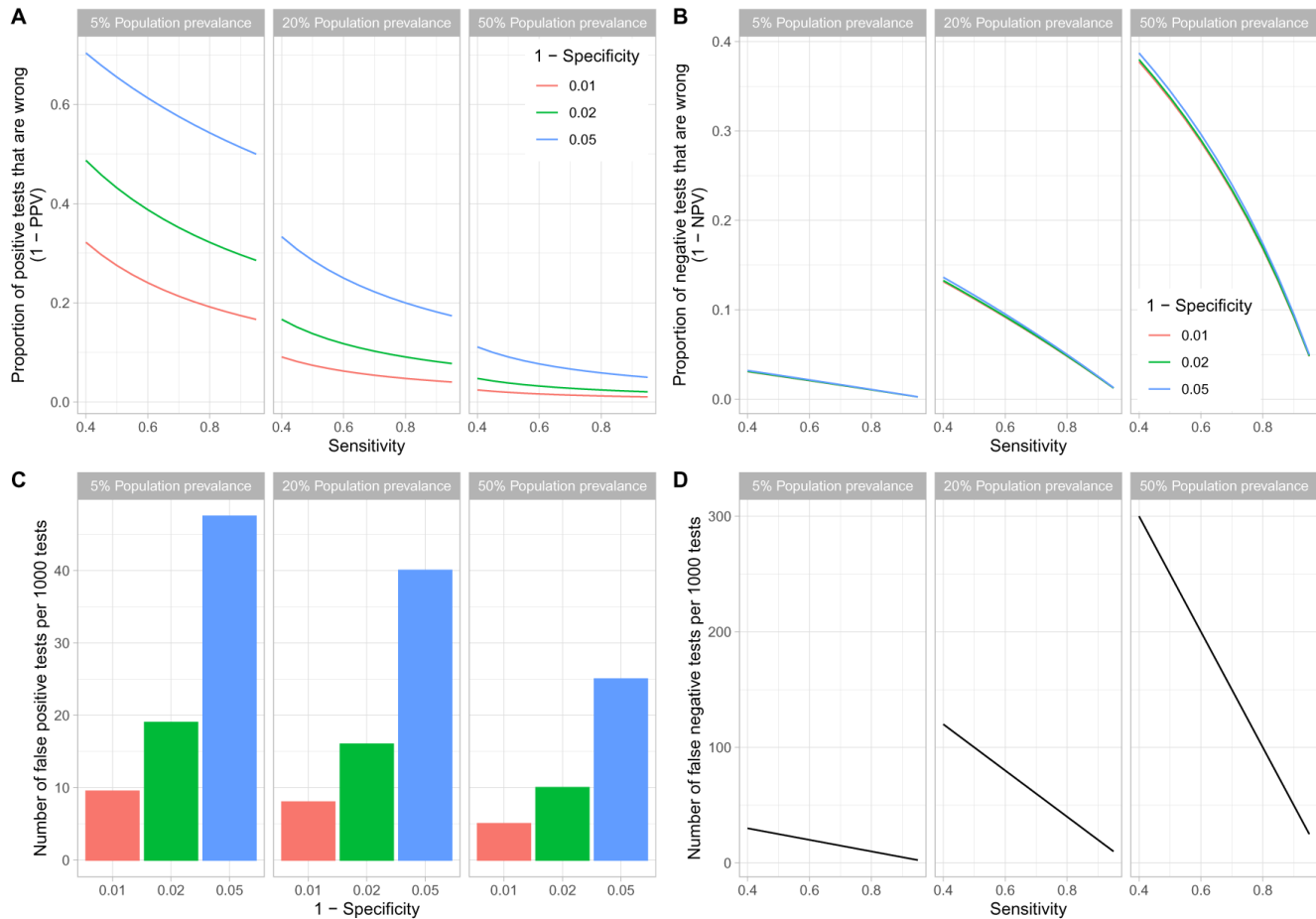


Figure 4. Influence of population prevalence of seropositivity on assay performance. Scenarios with population prevalence of 5%, 20% and 50% are shown within each panel. **(A)** The proportion of all positive tests that are wrong (1-positive predictive value), which would lead to false release from lock-down of non-immune individuals, for varying test sensitivity (x-axis) and 1-specificity (line colour). **(B)** The proportion of negative tests that are wrong. **(C)** The absolute number of false positive tests per 1000 tests. **(D)** The absolute number of false negative tests per 1000 tests.

Our study, and another undertaken independently in parallel¹¹, demonstrates accurate performance of ELISA targeting anti-spike protein antibodies. Additionally, our ELISA results are supported by context regarding disease severity and the time of sampling relative to symptom onset. Our data on the kinetics of antibody responses build upon studies of hospitalised patients in China reporting a median 11 days to seroconversion for total antibody, with IgM and IgG seroconversion at days 12 and 14, respectively¹⁸, and others that report 100% IgG positivity by 15–19 days^{19,23}. Our ELISA data show IgG titres rose over the first 3 weeks of infection and that IgM testing identified no additional cases. Methods to enhance sensitivity, especially shortly after symptom onset, could consider different sample types (e.g. saliva), different antibody classes (e.g. IgA)²⁴, T-cell assays or antigen detection²⁵. In contrast to others^{19,26–28}, we did not find evidence of an association between disease severity and antibody titres. We observed several LFIA false positives, which may have potentially resulted from cross-reactivity of non-specific antibodies (e.g. reflecting past exposure to other seasonal coronavirus infections).

The main study limitation is that numbers tested were too small to provide tight confidence intervals around performance estimates for any specific LFIA device. Expanding testing across diverse populations would increase certainty, but given the broadly comparable performance of different assays, the cost and manpower to test large numbers may not be justifiable. Demonstrating high specificity is particularly challenging; for example, if the true underlying value was 98%, 1000 negative controls would be required to estimate the specificity of an assay to $\pm 1\%$ with approximately 90% power. Full assessment should also include a range of geographical locations and ethnic groups, children, and those with immunological disease including autoimmune conditions and immunosuppression.

In summary, antibody testing is a crucial component of measures that may be required to inform release from lockdown. Our findings suggest that while current LFIA devices may provide some information for population-level surveys, their performance is inadequate for most individual patient applications. The ELISA we describe is currently being optimised

and adapted to run on a high-throughput platform and provides promise for the development of reliable approaches to antibody detection that can support decision making for clinicians, the public health community, policy-makers and industry.

Data availability

Underlying data

Figshare: Antibody testing for COVID-19: A report from the National COVID Scientific Advisory Panel [Supporting Data]. <https://doi.org/10.6084/m9.figshare.12229922>¹³.

This project contains the following underlying data:

- Supplementary_table_s1 (XLSX). (Metadata describing origin and characteristics of designated negative controls and individuals with confirmed SARS-CoV-2 infection.)
- Supplementary_table_s6 (XLSX). (Results of all assays performed and relevant metadata.)

Extended data

Figshare: Antibody testing for COVID-19: A report from the National COVID Scientific Advisory Panel [Supporting Data]. <https://doi.org/10.6084/m9.figshare.12229922>¹³.

File 'Supplementary material' (PDF) contains the following extended data:

- Supplementary methods.
- Figure S1. Sensitivity and specificity of lateral flow devices compared with RT-PCR confirmed cases and pre-pandemic controls (panels A and B) and compared with ELISA results (panels C and D).
- Figure S2. Comparison between ELISA and LFIA for SARS-CoV-2 designated negative and positive plasma.
- Supplementary table S2. Summary grid presenting the number of samples from each cohort tested using different assay platforms.
- Supplementary table S3. Multivariable regression models for relationship between ELISA IgM and IgG readings and covariates in RT-PCR positive cases.

- Supplementary table S4. Results of nine LFIA devices and an ELISA assay, tested with plasma classified as positive (RT-PCR positive) obtained from patients ≥ 10 days after onset of symptoms.
- Supplementary table S5. Results of nine LFIA devices, tested with plasma classified as positive and negative using ELISA as an alternative reference standard.

Reporting guidelines

Figshare: STARD checklist for 'Antibody testing for COVID-19: A report from the National COVID Scientific Advisory Panel'. <https://doi.org/10.6084/m9.figshare.12229922>¹³.

Data are available under the terms of the [Creative Commons Attribution 4.0 International license](#) (CC-BY 4.0).

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Author information

The members and affiliations of the National COVID Testing Scientific Advisory Panel are listed in the author list, in alphabetical order.

Author contributions

Leadership roles were undertaken by DC (project management), RJP and DJR (management of pre-pandemic negative sample bank), AJM (identification and recruitment of cases), DWE and PCM (oversight of LFIA, data analysis and manuscript writing), GRS (ELISA design, development and data generation).

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Muki Shey 

Wellcome Centre for Infectious Disease Research in Africa (CIDRI-Africa) & Department of Medicine, University of Cape Town, Cape Town, South Africa

The article by Adams and colleagues "Antibody testing for COVID-19: A report from the National COVID Scientific Advisory Panel" compares the performance of "in-house" ELISA with 9 lateral flow Immunoassays (LFIA) from different companies, using samples from pre-pandemic period and RT-PCR-confirmed COVID-19 cases at different stages of the disease. The ELISA showed 85% sensitivity and 100% specificity. The LFIA show higher sensitivity and specificity compared with ELISA than with RT-PCR, and the performance of the LFIA were similar to each other. The authors conclude that ELISA performs better than LFIA for individual patient applications.

Comments

1. The authors describe that the ELISA used measured antibodies against the spike protein but not much info is available about the different LFIA and what parts of the virus may have been used to develop and optimize the antibody detection. This information is important to have a complete understanding of the comparison between the ELISA and the LFIA (Table 1).
2. Most recommended quarantine/isolation period (may be country-dependent) is 10-14 days from symptom onset and the authors describe up to 28 days as acute period. A justification may be necessary. Was this predefined, based on information around viral shedding or antibody kinetics (similar to data presented in Figure 2D)?
3. In line with Tables 1 and S4 and the false negative results, can the authors comment on the antibodies possibly targeting other parts of the virus other than spike proteins (especially for the ELISA since details of the LFIA are not available)?

Is the work clearly and accurately presented and does it cite the current literature?

Yes

Is the study design appropriate and is the work technically sound?

Yes

Are sufficient details of methods and analysis provided to allow replication by others?

Yes

If applicable, is the statistical analysis and its interpretation appropriate?

Yes

Are all the source data underlying the results available to ensure full reproducibility?

Partly

Are the conclusions drawn adequately supported by the results?

Yes

Competing Interests: No competing interests were disclosed.

Reviewer Expertise: Immunology and infectious Diseases.

I confirm that I have read this submission and believe that I have an appropriate level of expertise to confirm that it is of an acceptable scientific standard.

Reviewer Report 15 September 2020

<https://doi.org/10.21956/wellcomeopenres.17469.r40226>

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Kim Tien Ng 

Infectious Disease programme, Department of Microbiology and Immunology, National University Health System (NUHS), National University of Singapore, Singapore, Singapore

Yee Joo Tan 

Infectious Disease programme, Department of Microbiology and Immunology, National University Health System (NUHS), National University of Singapore & Institute of Molecular and Cell Biology (IMCB), A*STAR (Agency for Science, Technology and Research), Singapore, Singapore

This is a systematic study that compares a laboratory-based home-made ELISA serological assay for COVID-19 with nine commercially-available rapid lateral flow immunoassay (LFIA). The results presented are convincingly and important for the development/improvement of serological assays for COVID-19. It will be helpful if the authors can clarify the following points:

1. Page 4, Methods: 2nd paragraph, line 4: Acute (28 days).
Query: How did the authors decide on the 28 days cut-off? Is this definition similar to other studies on COVID-19 seroconversion? It would be good to discuss and include citation.

2. Page 6, it was stated "We also considered performance relative to ELISA (*Extended data*, Table S5, Figure S1), because the LFIA devices target the same antibodies."
Query: The viral protein used each of the LFIA device is not stated in the manuscript. Based on this sentence, are the authors stating that all of them use SARS-CoV-2 Spike residues 1-1208? If not, please state the viral protein or fragment used for each device.
3. Page 9, "... with animal data demonstrating that SARS-CoV-2 anti-spike IgG contributes to a proinflammatory response associated with lung injury in macaques."
Query: SARS-CoV-2 should be SARS-CoV.
4. Page 10, it was stated "We observed several LFIA false positives, which may have potentially resulted from cross-reactivity of non-specific antibodies (e.g. reflecting past exposure to other seasonal coronavirus infections).
Query: Are these LFIA using the same viral protein or fragment? What is the sequence homology between SARS-CoV-2 and endemic coronaviruses for each of viral protein used?
5. Page 10, it was stated "In contrast to others, we did not find evidence of an association between disease severity and antibody titres"
Query: Is there any difference in study design between this study and the published studies cited? Were the published studies measuring anti-Spike binding antibodies or were they measuring neutralizing antibodies? Are the sample size and distribution of cases (mild vs severe, etc) comparable? Please highlight any limitation that may explain the discrepancy.
6. For the simulations shown in Figure 4.
Query: Please provide details on method/program as well as assumptions used.
7. The ELISA described by the authors is important and adds value to the development of serological assay for COVID-19. To briefly show the robustness of the assay, can authors provide information on (a) intra-assay variability, (b) inter-assay variability, (c) inter-operators variations and (d) the turnaround time i.e. from sample to result?

Is the work clearly and accurately presented and does it cite the current literature?

Partly

Is the study design appropriate and is the work technically sound?

Yes

Are sufficient details of methods and analysis provided to allow replication by others?

Partly

If applicable, is the statistical analysis and its interpretation appropriate?

Yes

Are all the source data underlying the results available to ensure full reproducibility?

Yes

Are the conclusions drawn adequately supported by the results?

Yes

Competing Interests: No competing interests were disclosed.

Reviewer Expertise: Molecular virology; Monoclonal antibody technology;

We confirm that we have read this submission and believe that we have an appropriate level of expertise to confirm that it is of an acceptable scientific standard.
